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Impact of one-carbon metabolism-related gene polymorphisms on risk of lung cancer in Japan: a case–control study

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There is substantial evidence that the decreased risk of lung cancer with high intake of vegetables and fruits is linked to folate as a specific nutrient. Functional polymorphisms in genes encoding one-carbon metabolism enzymes, methylenetetrahydrofolate reductase (*MTHFR* C677T and A1298C), methionine synthase (*MTR* A2756G), methionine synthase reductase (*MTRR* A66G) and thymidylate synthase, influence folate metabolism and thus might be suspected of impacting on lung cancer risk. We therefore conducted a case–control study with 515 lung cancer cases newly and histologically diagnosed and 1030 age- and sex-matched non-cancer controls to clarify associations with these five polymorphisms according to lung cancer subtype. Gene–environment interactions with smoking and drinking habit and folate consumption were also evaluated by logistic regression analysis. None of the polymorphisms showed any significant impact on lung cancer overall risk by genotype alone, but on histology-based analysis increase in *MTHFR* 677T and 1298C alleles was associated with reduced risk of squamous/small cell carcinoma ($P = 0.029$), especially among heavy smokers ($P = 0.035$), whereas the *MTHFR* 677TT genotype was linked to decreased risk for these subtypes among heavy drinkers (odds ratio = 0.17, 95% confidence interval: 0.03–0.98). In addition, we found interactions between the *MTRR* A66G polymorphism and smoking ($P = 0.015$) and the *MTHFR* A1298C polymorphism and alcohol consumption ($P = 0.025$) for risk of lung cancer overall. In conclusion, the results suggest that *MTHFR* polymorphisms contribute to risk of squamous/small cell carcinomas of the lung, along with possible interactions among folate metabolism-related polymorphisms and smoking/drinking habits. Further evaluation is warranted.

Introduction

Lung cancer, with its four major histological types (adenocarcinoma, squamous cell carcinoma, large cell carcinoma and small cell carcinoma), currently claims >55 000 lives annually in Japan and has become the leading cause of cancer death (1). Despite rapid advances in treatment over recent decades, the prognosis has not greatly improved. Therefore, efforts toward primary prevention in addition to early detection have come under the spotlight.

Abbreviations: CI, confidence interval; FFQ, food frequency questionnaire; 5,10-methylene THF, 5,10-methylenetetrahydrofolate; MTHFR, methylenetetrahydrofolate reductase; MTR, methionine synthase; MTRR, methionine synthase reductase; OR, odds ratio; PCR, polymerase chain reaction; 2R, two repeat; TS, thymidylate synthase; VNTR, variable number of tandem repeat.

Many epidemiological studies have provided evidence that high consumption of vegetables and fruits is associated with a reduced risk of lung cancer (2–4). Folate is one of the constituents found in vegetables and fruits, and dietary folate may be one of the micronutrients that provide protection against lung carcinogenesis (5–7).

Biological functions of folate within so-called 'one-carbon metabolism' are to facilitate *de novo* deoxynucleoside triphosphate synthesis and to provide methyl groups required for intracellular methylation reactions. Folate deficiency is thought to increase the risk of cancer through impaired DNA repair synthesis and disruption of DNA methylation that may lead to proto-oncogene activation (8–10).

Methylenetetrahydrofolate reductase (MTHFR), methionine synthase (MTR), methionine synthase reductase (MTRR) and thymidylate synthase (TS) play important and interrelated roles in folate metabolism (Figure 1). The MTHFR reduces 5,10-methylenetetrahydrofolate (5,10-methylene THF) to 5-methyl THF, the primary circulating form of folate (11). The TS catalyzes the conversion of deoxyuridine monophosphate to deoxythymidine monophosphate using 5,10-methylene THF (12). The MTHFR product, 5-methyl THF, is the methyl group donor for the remethylation of homocysteine to methionine catalyzed by MTR (13). MTR activity is maintained by MTRR (14). Polymorphisms in the genes for *MTHFR* C677T and A1298C, *MTR* A2756G, *MTRR* A66G and *TS* 28 bp variable number of tandem repeat (VNTR) in the promoter region are known to have functional relevance (15). Thus, they might play roles in the etiology of lung cancer in combination with environmental factors such as folate consumption. Since information for this area of lung cancer is limited (16–22), we conducted the present case–control study, taking tobacco smoking, alcohol drinking and intake of folate into consideration.

Materials and methods

Subjects

The cases were 515 patients who were newly and histologically diagnosed as having lung cancer and not having any earlier history of cancer. Controls ($n = 1030$) were randomly selected and matched by age (±3 years) and sex to cases with a 1:2 case–control ratio from among the 2395 cancer-free individuals. All the subjects were recruited in the framework of the Hospital-based Epidemiologic Research Program at Aichi Cancer Center, as described elsewhere (23,24). In brief, information on lifestyle factors was collected using a self-administered questionnaire, checked by a trained interviewer, from all first-visit out-patients at Aichi Cancer Center Hospital aged 18–79 who were enrolled in Hospital-based Epidemiologic Research Program at Aichi Cancer Center between January 2001 and November 2005. Out-patients were also asked to provide blood samples. Each patient was asked about his or her lifestyle when healthy or before the current symptoms developed. Approximately 95% of eligible subjects complete the questionnaire and 60% provide blood samples. The data were loaded into a Hospital-based Epidemiologic Research Program at Aichi Cancer Center database and routinely linked with the hospital-based cancer registry system to update the data on cancer incidence. All participants gave written informed consent and the study was approved by Institutional Ethical Committee of Aichi Cancer Center.

Genotyping of MTHFR, MTR, MTRR and TS

DNA from each subject was extracted from the buffy coat fraction using BioRobot EZ1 and an EZ1 DNA Blood 350 ml Kit (Qiagen, Tokyo, Japan). The genotyping method was described in our previous reports with the polymerase chain reaction (PCR) TaqMan method using the GeneAmp PCR System 9700 or the 7500 Fast Real-Time PCR system (Applied Biosystems, Foster City, CA). Briefly, for the *MTHFR* C677T (dbSNP ID: rs677) and A1298C (rs1801131), as well as *MTR* A2756G (rs1805087) and *MTRR* A66G (rs1801394) polymorphisms, extracted DNA was amplified with validated probes (assay IDs: C__11975651_10, C__850486_20, C__12005959_10 and C__3068176_10, respectively; Applied Biosystems). The *TS* VNTR polymorphism was defined by PCR using 5′-CGTGGCTCCTGCGTTTCC-3′ and 5′-GAGCCGGCCACAGGCAT-3′ primers. In our laboratory, quality of genotyping is routinely assessed statistically using the Hardy–Weinberg test.

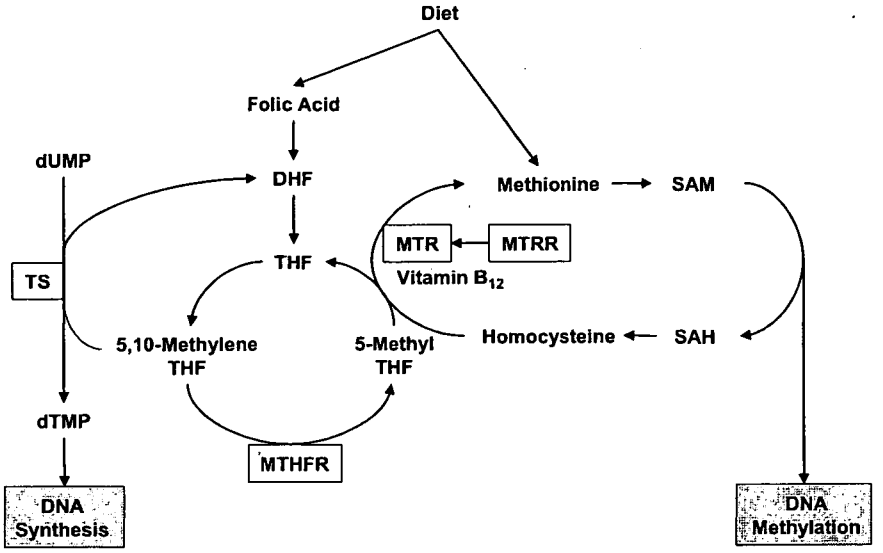

Fig. 1. Overview of folate metabolism. Enzymes with polymorphisms investigated in this study are boxed. THF, tetrahydrofolate; DHF, dihydrofolate; dUMP, deoxyuridine monophosphate; dTMP, deoxythymidine monophosphate; SAM, *S*-adenosylmethionine and SAH, *S*-adenosylhomocysteine.

When allelic distributions for controls depart from the Hardy–Weinberg frequency, genotyping is assessed using another method.

Intake assessment for folate and other nutrients

The consumption of folate and other nutrients was determined using a food frequency questionnaire (FFQ), described in detail elsewhere (25,26). Briefly, the FFQ consisted of 47 single food items with frequencies in the eight categories. We estimated the average daily intake of nutrients by multiplying the food intake (in grams) or serving size by the nutrient content per 100 g of food as listed in standard tables of food composition. Consumption of folate and other vitamins from supplements was not considered in total consumption because the questionnaire for multivitamins was not quantitative. Energy-adjusted intake of nutrients was calculated by the residual method (27). The FFQ was validated by referring to a 3-day weighed dietary record as a standard, which showed reproducibility and validity to be acceptable (28). The deattenuated correlation coefficients for energy-adjusted intakes of folate were 0.36 in men and 0.38 in women.

Consumption of tobacco and alcohol

Cumulative smoking dose was evaluated as pack-years, the product of the number of packs consumed per day and years of smoking. Smoking habit was entered for four categories of never, former and current smokers of <40 and ≥40 pack-years. Former smokers were defined as those who quit smoking at least 1 year before the survey. Consumption of each type of beverage (Japanese sake, beer, shochu, whiskey and wine) was determined by the average number of drinks per day, which was then converted into a Japanese sake (rice wine) equivalent. One drink equates to one 'go' (180 ml) of Japanese sake, which contains 23 g of ethanol, equivalent to one large bottle (633 ml) of beer, two shots (60 ml) of whiskey and two and a half glasses of wine (200 ml). One drink of 'shochu' (distilled spirit), which contains 25% ethanol, was rated as 108 ml. Total amount of alcohol consumption was estimated as the summarized amount of pure alcohol consumption (gram per drink) of Japanese sake, beer, shochu, whiskey and wine among current regular drinkers. Drinking habit was entered for four categories of never, former, current moderate and heavy drinkers. Heavy drinkers were defined as those currently drinking alcoholic beverages 5 days or more per week in a daily amount of 46 g (two Japanese drinks) or more, whereas moderate drinkers were defined as those currently consuming less frequently than 5 days/week, in lower amounts, or both. Former drinkers were defined as those who quit drinking at least 1 year before the survey. Former or current smokers and drinkers were categorized as 'smokers' and 'drinkers', respectively.

Statistical analysis

To assess the strength of the associations between polymorphic genes involved in folate metabolism and risk of lung cancer, odds ratio (ORs) with 95% confidence intervals (CIs) were estimated using age- and sex-matched conditional logistic models adjusted for potential confounders. For stratified and interaction analysis by smoking and drinking habit and folate intake, an unconditional logistic regression model was used because the matching was not retained after stratification by smoking and drinking habit and folate intake. Folate and other nutrient intakes were categorized into three groups as: first, second and third tertiles of dietary intake among controls. Potential confounders considered in the multivariate analyses were age, sex, smoking habit (never smokers, former smokers, current smokers of <40 or ≥40 pack-years), drinking habit (never drinkers, former drinkers, moderate drinkers or heavy drinkers), body mass index (<18.5, 18.5–24.9 or ≥25.0), total energy intake (as a continuous variable), dietary carotene intake (μg/day, tertiles), dietary vitamin C intake (mg/day, tertiles), dietary vitamin E intake (mg/day, tertiles), dietary folate intake (μg/day, tertiles), multivitamin use (at least once per week for 1 year or longer: yes or no) and referral pattern (patient's discretion, family recommendation, referral from other clinics, secondary screening after primary screening or others). Missing values for each covariate were treated as an additional category in the variable and were included in the logistic model.

For the histology-based analysis, we combined squamous cell carcinoma and small cell carcinoma, because tumors of these subtypes were small in number and both are consistently more related with smoking as compared with adenocarcinomas. Considering potential effects of two polymorphisms (*MTHFR* C677T and *MTHFR* A1298C) on lung risk, we evaluated associations with their combined genotypes. Trend of genotype was assessed by score test applying score for each genotype (0, homozygous for reference allele or combined reference genotypes; 1, heterozygote or one reference genotype and 2, homozygous non-reference allele or non-reference genotype).

Gene–environment interactions between smoking and drinking habit and folate intake and genotypes in each polymorphism were evaluated under the multiplicative assumption. Products of scores for genotype (0, homozygous; 1, heterozygote and 2, homozygous or 0, referent alleles and 1, non-referent alleles) and smoking habit (0, non-smoker and 1, smoker), drinking habit (0, non-drinker and 1, drinker), folate intake (0, tertile 1 and 1, tertile 2 + 3) or combined smoking–drinking habit (0, non-smoker and non-drinker; 1, smoker and non-drinker or drinker and non-smoker and 2, smoker/drinker) were included as interaction terms. Differences in categorized demographic variables between the cases and controls were tested by the Chi-squared test. Mean values for age and total energy intake were compared for cases and controls by the Student's *t*-test. Accordance with the Hardy–Weinberg equilibrium was checked for controls using the Chi-squared test and the exact *P*-value was used to assess any discrepancies between genotypes and allele frequencies. A *P*-value <0.05 was considered statistically significant. All analyses were performed using STATA version 9 (Stata Corp., College Station, TX).

Results

Data from 515 lung cancer cases, comprising 316 (61.4%) adenocarcinomas, 91 (17.7%) squamous cell carcinomas, 55 (10.7%) small

cell carcinomas, 40 (7.8%) large cell carcinomas and 13 (2.5%) others, and 1030 controls were available for analysis. Table I shows the distribution of cases and controls by background characteristics. Age and sex were appropriately matched. Smoking habits differed to a large extent between cases and controls. The proportion of 40 pack-years or more current smokers in cases was significantly higher than controls. Heavy drinkers in the cases were significantly higher than for the controls. Among cases, the proportion of lower body mass index was higher, consistent with previous study (29). Total energy intake did not differ between cases and controls. Significant lower intake of dietary carotene was found among the cases. For other nutrients lower proportions of the highest intake group among the cases also were found, including for folate, but these were not statistically significant. With regard to referral pattern, referral from other clinics was frequent, whereas patient discretion and secondary screening after primary screening were less common among the case group than the control group.

Table II shows genotype distributions for *MTHFR*, *MTR*, *MTRR* and *TS* and their ORs and 95% CIs for lung cancer risk according to histological subtypes. The genotype frequencies for all the polymorphisms were in accordance with the Hardy–Weinberg law in controls: *MTHFR* C677T (P = 0.17), *MTHFR* A1298C (P = 0.51), *MTR* A2756G (P = 0.17), *MTRR* A66G (P = 0.85) and *TS* VNTR (P = 0.51). On analysis of lung cancer overall, a slightly reduced risk was observed with the *MTHFR* 677TT genotype, but without statistical significance. The genotype frequencies for *TS* VNTR were quite varied; however, two repeat (2R) and three repeat alleles were dominant. The 2R/2R genotype showed decreased risk of lung cancer as compared with the non-2R homozygous, although again this was not significant. On subanalysis according to histological subtypes, the combination of *MTHFR* C677T and A1298C polymorphisms showed a significant decreased risk of squamous/small cell carcinoma among individuals with two or more *MTHFR* 677T and/or 1298C alleles (OR = 0.34, 95% CI: 0.13–0.92, trend P = 0.029), compared with those with *MTHFR* 677CC and 1298AA genotypes. In contrast, none of the polymorphisms showed any significant impact on adenocarcinoma risk.

To further evaluate the impact of *MTHFR* polymorphisms with regard to squamous/small cell carcinoma, we conducted stratified analysis by smoking and drinking habit (Table III). Among heavy drinkers, the *MTHFR* 677TT genotype conferred a significant decreased risk (OR = 0.17, 95% CI: 0.03–0.98, trend P = 0.041). A significant decreased risk among 40 pack-years or more current smokers was observed as number of *MTHFR* 677T or 1298C alleles increased (trend P = 0.035). No clear association was found for lung cancers overall or for adenocarcinomas in the stratified analysis (data not shown).

Table IV shows data for the combinations of gene and environmental factors with reference to lung cancer overall risk. The interaction with smoking was significant for the *MTRR* A66G genotype (P = 0.015). Among non-smokers, risk was reduced with increase in the number of *MTRR* G alleles, whereas a trend for increased risk was observed among smokers. A significant interaction between drinking habits and the *MTHFR* A1298C genotype was found (P = 0.025). These two interactions were especially noteworthy for adenocarcinomas when histology-based analyses were conducted (data not shown). We were not able to analyze the smoking interaction for squamous/small cell due to insufficient number of non-smokers in this category. No obvious interaction was found between folate intake and the polymorphisms.

Considering the possible effects of both tobacco smoking and alcohol drinking on folate, we further examined the impact of four-way combinations of these two factors, folate intake and the polymorphisms on lung cancer risk (Table V). The *MTRR* A66G genotype showed a significant interaction among the subjects with tertiles 2 or 3 of folate intake (P = 0.023). The risk with the *MTRR* 66GG was consistently decreased among non-smoker/non-drinker subjects with adequate folate intake (OR = 0.20, 95% CI: 0.04–0.91).

Table I. Characteristics of cases and controls

	Cases (n = 515), n(%)	Controls (n = 1030), n(%)	P-value
Age			
<50	53 (10.3)	108 (10.5)	
50–59	142 (27.6)	283 (27.5)	
60–69	193 (37.5)	389 (37.8)	
70–79	127 (24.7)	250 (24.3)	1.00
Mean age ± SD	61.9 ± 9.9	61.8 ± 9.8	0.87
Sex			
Male	381 (74.0)	762 (74.0)	
Female	134 (26.0)	268 (26.0)	1.00
Smoking status			
Never	129 (25.0)	401 (38.9)	
Former[a]	111 (21.6)	310 (30.1)	
Current (pack-years)			
0–39	71 (13.8)	149 (14.5)	
≥40	197 (38.3)	161 (15.6)	<0.01
Unknown	7 (1.4)	9 (0.9)	
Drinking status			
Never	196 (38.1)	378 (36.7)	
Former[a]	15 (2.9)	56 (5.4)	
Current			
Moderate[b]	192 (37.3)	454 (44.1)	
Heavy[c]	98 (19.0)	119 (11.6)	<0.01
Unknown	14 (2.7)	23 (2.2)	
BMI			
<18.5	38 (7.4)	55 (5.3)	
18.5–24.9	381 (74.0)	720 (69.9)	
≥25.0	94 (18.3)	249 (24.2)	0.03
Unknown	2 (0.4)	6 (0.6)	
Mean total energy ± SD, kcal/day	1670 ± 372	1677 ± 352	0.73
Carotene (μg/day)			
Tertile 1 (1331.2–2305.9)	200 (38.8)	341 (33.1)	
Tertile 2 (2306.0–3312.6)	149 (28.9)	341 (33.1)	
Tertile 3 (3312.7–12801.4)	158 (30.7)	341 (33.1)	0.04
Unknown	8 (1.6)	7 (0.7)	
Vitamin C (mg/day)			
Tertile 1 (26.8–74.5)	188 (36.5)	342 (33.2)	
Tertile 2 (74.6–102.0)	161 (31.3)	342 (33.2)	
Tertile 3 (102.1–364.5)	159 (30.7)	341 (33.1)	0.15
Unknown	7 (1.4)	5 (0.5)	
Vitamin E (total α-mg/day)			
Tertile 1 (1.5–4.8)	193 (37.5)	342 (33.2)	
Tertile 2 (4.9–6.3)	168 (32.6)	342 (33.2)	
Tertile 3 (6.4–17.1)	151 (29.3)	342 (33.2)	0.29
Unknown	3 (0.6)	4 (0.4)	
Folate intake (μg/day)			
Tertile 1 (139.5–274.5)	191 (37.1)	342 (33.2)	
Tertile 2 (274.6–354.9)	156 (30.3)	342 (33.2)	
Tertile 3 (355.0–1481.0)	162 (31.5)	341 (33.1)	0.18
Unknown	6 (1.2)	5 (0.5)	
Multivitamin use (at least once per week for 1 year or longer)			
Yes	111 (21.6)	253 (24.6)	
No	380 (73.8)	721 (70.0)	0.30
Unknown	24 (4.7)	56 (5.4)	
Referral pattern to our hospital			
Patient's discretion	52 (10.1)	306 (29.7)	
Family recommendation	86 (16.7)	195 (18.9)	
Referral from other clinics	287 (55.7)	300 (29.1)	
Secondary screening after primary screening	83 (16.1)	214 (20.8)	
Others	2 (0.4)	10 (1.0)	<0.01
Unknown	5 (1.0)	5 (0.5)	

SD: standard deviation, BMI: body mass index.
[a]Former smokers and drinkers were defined as subjects who had quit smoking and drinking at least 1 year previously.
[b]Moderate drinker means <46 g ethanol/drink and/or <5 days/week.
[c]Heavy drinker means ≥46 g ethanol/drink and ≥5 days/week.

Table II. *MTHFR, MTR, MTRR* and *TS* genotype distributions, and ORs for lung cancer according to histology

	All			Adenocarcinoma			Squamous + small cell carcinoma		
	Cases (n = 515), n (%)	Controls (n = 1030), n (%)	ORs[a] (95% CIs)	Cases (n = 316), n (%)	Controls (n = 632), n (%)	ORs[a] (95% CIs)	Cases (n = 146), n (%)	Controls (n = 292), n (%)	ORs[a] (95% CIs)
MTHFR (C677T)									
CC	182 (35.3)	379 (36.8)	1.00 (ref.)	109 (34.5)	237 (37.5)	1.00 (ref.)	54 (37.0)	103 (35.3)	1.00 (ref.)
CT	256 (49.7)	474 (46.0)	1.05 (0.81–1.37)	158 (50.0)	288 (45.6)	1.01 (0.72–1.41)	72 (49.3)	134 (45.9)	0.83 (0.43–1.58)
TT	77 (15.0)	177 (17.2)	0.75 (0.52–1.09)	49 (15.5)	107 (16.9)	0.85 (0.53–1.34)	20 (13.7)	55 (18.8)	0.44 (0.16–1.18)
P_{trend}[b]			0.260			0.567			0.129
MTHFR (A1298C)									
AA	341 (66.2)	652 (63.3)	1.00 (ref.)	210 (66.5)	416 (65.8)	1.00 (ref.)	94 (64.4)	175 (59.9)	1.00 (ref.)
AC	149 (28.9)	322 (31.3)	0.85 (0.65–1.13)	90 (28.5)	189 (29.9)	0.94 (0.67–1.33)	46 (31.5)	99 (33.9)	0.84 (0.42–1.68)
CC	22 (4.3)	45 (4.4)	1.01 (0.56–1.83)	14 (4.4)	22 (3.5)	1.46 (0.68–3.16)	5 (3.4)	14 (4.8)	0.40 (0.07–2.28)
UK[c]	3 (0.6)	11 (1.1)		2 (0.6)	5 (0.8)		1 (0.7)	4 (1.4)	
P_{trend}[b]			0.428			0.822			0.348
MTHFR C677T and A1298C combined Number of Variants									
0	76 (14.8)	174 (16.9)	1.00 (ref.)	43 (13.6)	118 (18.7)	1.00 (ref.)	23 (15.8)	41 (14.0)	1.00 (ref.)
1	273 (53.0)	471 (45.7)	1.19 (0.83–1.71)	171 (54.1)	293 (46.4)	1.46 (0.93–2.27)	76 (52.1)	130 (44.5)	0.52 (0.19–1.40)
≦2	163 (31.7)	374 (36.3)	0.84 (0.58–1.24)	100 (31.6)	216 (34.2)	1.10 (0.68–1.77)	46 (31.5)	117 (40.1)	0.34 (0.13–0.92)
UK[c]	3 (0.6)	11 (1.1)		2 (0.6)	5 (0.8)		1 (0.7)	4 (1.4)	
P_{trend}[b]			0.110			0.819			0.029
MTR (A2756G)									
AA	319 (61.9)	698 (67.8)	1.00 (ref.)	192 (60.8)	423 (66.9)	1.00 (ref.)	100 (68.5)	195 (66.8)	1.00 (ref.)
AG	175 (34.0)	291 (28.3)	1.23 (0.94–1.60)	109 (34.5)	184 (29.1)	1.26 (0.91–1.75)	42 (28.8)	84 (28.8)	0.80 (0.42–1.52)
GG	21 (4.1)	40 (3.9)	1.04 (0.55–2.00)	15 (4.7)	25 (4.0)	1.35 (0.62–2.91)	4 (2.7)	13 (4.5)	0.49 (0.07–3.38)
UK[c]	0 (0)	1 (0.1)							
P_{trend}[b]			0.227			0.146			0.364
MTRR (A66G)									
AA	235 (45.6)	484 (47.0)	1.00 (ref.)	148 (46.8)	294 (46.5)	1.00 (ref.)	63 (43.2)	136 (46.6)	1.00 (ref.)
AG	226 (43.9)	446 (43.3)	1.02 (0.79–1.31)	139 (44.0)	275 (43.5)	0.93 (068–1.28)	64 (43.8)	131 (44.9)	1.18 (0.60–2.31)
GG	54 (10.5)	100 (9.7)	0.96 (0.62–1.47)	29 (9.2)	63 (10.0)	0.91 (0.52–1.58)	19 (13.0)	25 (8.6)	1.11 (0.38–3.21)
P_{trend}[b]			0.939			0.638			0.718
TS VNTR									
Non-2R/non-2R	372 (72.2)	721 (70.0)	1.00 (ref.)	236 (74.7)	434 (68.7)	1.00 (ref.)	101 (69.2)	212 (72.6)	1.00 (ref.)
2R/non-2R	132 (25.6)	278 (27.0)	0.96 (0.73–1.27)	73 (23.1)	181 (28.6)	0.81 (0.57–1.13)	43 (29.5)	69 (23.6)	1.26 (0.61–2.59)
2R/2R	10 (1.9)	31 (3.0)	0.63 (0.29–1.39)	6 (1.9)	17 (2.7)	0.62 (0.22–1.73)	2 (1.4)	11 (3.8)	0.23 (0.03–1.62)
UK[c]	1 (0.2)	0 (0)		1 (0.3)	0 (0)				
P_{trend}[b]			0.394			0.137			0.653

[a]ORs were matched for age and sex and adjusted for smoking habit, drinking habit, body mass index, total energy intake, carotene intake, vitamin C intake, vitamin E intake, multivitamin use and referral pattern to our hospital.

[b]Trend of genotype was assessed by score test applying score for each genotypes (0, homozygous for reference allele or combined reference genotypes; 1, heterozygote or one reference genotype and 2, homozygous non-reference allele or non-reference genotype).

[c]UK denotes genotype unknown.

Table III. Stratification analysis by smoking and drinking habit for the *MTHFR* polymorphisms in squamous/small cell carcinoma

	Smoking status						Drinking status							
	Smokers		0–39 pack-years		40 ≥ pack-years		Non-drinkers		Drinkers		Moderate drinkers		Heavy drinkers	
	Cases/ controls	ORs[a] (95% CIs)	Cases/ controls	ORs[a] (95% CIs)	Cases/ controls	ORs[a] (95% CIs)	Cases/ controls	ORs[a] (95% CIs)	Cases/ controls	ORs[a] (95% CIs)	Cases/ controls	ORs[a] (95% CIs)	Cases/ controls	ORs[a] (95% CIs)
MTHFR (C677T)														
CC	53/229	1.00 (ref.)	7/65	1.00 (ref.)	35/58	1.00 (ref.)	15/143	1.00 (ref.)	39/236	1.00 (ref.)	22/168	1.00 (ref.)	14/38	1.00 (ref.)
CT	72/293	0.94 (0.61–1.45)	10/65	1.83 (0.40–8.47)	41/73	0.85 (0.43–1.67)	23/176	0.89 (0.36–2.21)	49/298	1.08 (0.63–1.84)	26/206	1.02 (0.48–2.16)	14/61	0.37 (0.09–1.63)
TT	20/107	0.81 (0.44–1.51)	4/19	5.63 (0.60–53.25)	11/30	0.50 (0.20–1.29)	3/59	0.52 (0.09–3.00)	17/118	0.90 (0.44–1.84)	8/80	0.85 (0.30–2.43)	5/20	0.17 (0.03–0.98)
P_{trend}[b]		0.528		0.151		0.179		0.519		0.871		0.817		0.041
MTHFR (A1298C)														
AA	93/406	1.00 (ref.)	15/106	1.00 (ref.)	57/97	1.00 (ref.)	29/232	1.00 (ref.)	65/420	1.00 (ref.)	36/293	1.00 (ref.)	18/74	1.00 (ref.)
AC	46/186	1.04 (0.68–1.61)	5/33	0.84 (0.15–4.63)	26/55	0.68 (0.35–1.33)	11/123	0.67 (0.26–1.77)	35/199	1.17 (0.69–2.01)	18/139	0.99 (0.46–2.11)	13/40	3.61 (0.87–14.96)
CC	5/30	0.80 (0.28–2.26)	0/8	NA[d]	4/9	0.84 (0.21–3.31)	1/20	0.30 (0.02–3.82)	4/25	1.05 (0.31–3.51)	2/17	1.03 (0.19–5.74)	2/5	1.05 (0.08–13.61)
UK[c]	1/7		1/2				0/3		1/8		0/5			
P_{trend}[b]		0.916		0.381		0.362		0.263		0.649		0.998		0.300
MTHFR C677T and A1298C combined Number of Variants														
0	22/105	1.00 (ref.)	3/37	1.00 (ref.)	15/22	1.00 (ref.)	6/64	1.00 (ref.)	17/110	1.00 (ref.)	13/81	1.00 (ref.)	4/15	1.00 (ref.)
1	76/295	1.08 (0.61–1.92)	11/72	0.63 (0.10–3.98)	47/73	0.97 (0.39–2.43)	28/169	1.02 (0.30–3.50)	48/302	1.13 (0.57–2.27)	22/211	0.57 (0.23–1.44)	17/58	0.93 (0.12–6.96)
≦2	46/222	0.88 (0.48–1.63)	6/38	2.40 (0.30–18.97)	25/66	0.44 (0.17–1.17)	7/142	036 (0.08–1.56)	39/232	1.15 (0.56–2.36)	21/157	0.82 (0.32–2.13)	12/46	0.30 (0.04–2.55)
UK[c]	1/7		1/2				0/3		1/8		0/5			
P_{trend}[b]		0.557		0.397		0.035		0.109		0.751		0.860		0.148

Data were not available in non-smokers because of absence of subjects in this category.

[a]ORs were adjusted for age, sex, smoking habit, drinking habit, body mass index, total energy intake, carotene intake, vitamin C intake, vitamin E intake, folate intake, multivitamin use and referral pattern to our hospital.

[b]Trend of genotype was assessed by score test applying score for each genotypes (0, homozygous for reference allele; 1, heterozygote and 2, homozygous non-reference allele).

[c]UK denotes genotype unknown.

[d]NA indicates not available because of absence of subjects in this category.

Table IV. Interaction between *MTHFR, MTR, MTRR* and *TS* polymorphisms and smoking and drinking habit and folate intake for lung cancer risk

	Smoking habit			Drinking habit			Folate intake		
	Non-smoker	Smoker	*P* interaction[b]	Non-drinker	Drinker	*P* interaction[b]	Tertile 1 (139.5–274.5 μg/day)	Tertile 2 + 3 (274.6–1481.0 μg/day)	*P* interaction[b]
	ORs[a] (95% CIs)	ORs[a] (95% CIs)		ORs[a] (95% CIs)	ORs[a] (95% CIs)		ORs[a] (95% CIs)	ORs[a] (95% CIs)	
MTHFR (C677T)									
CC	1.00 (ref.)	2.59 (1.61–4.17)		1.00 (ref.)	1.02 (0.67–1.55)		1.00 (ref.)	0.82 (1.34–0.82)	
CT	1.09 (0.69–1.73)	2.84 (1.79–4.52)		1.17 (0.77–1.78)	1.07 (0.72–1.60)		1.03 (1.58–1.03)	0.93 (1.50–0.93)	
TT	0.68 (0.36–1.29)	2.55 (1.48–4.40)	0.430	1.21 (0.68–2.14)	0.74 (0.45–1.23)	0.207	0.98 (1.82–0.98)	0.69 (1.22–0.69)	0.851
MTHFR (A1298C)									
AA	1.00 (ref.)	2.80 (1.88–4.18)		1.00 (ref.)	0.76 (0.56–1.04)		1.00 (ref.)	0.81 (1.24–0.81)	
AC	0.88 (0.55–1.39)	2.61 (1.67–4.07)		0.73 (0.48–1.12)	0.77 (0.53–1.11)		0.73 (1.14–0.73)	0.80 (1.27–0.80)	
CC	1.60 (0.61–4.19)	1.84 (0.85–3.96)	0.464	0.36 (0.12–1.04)	1.20 (0.58–2.47)	0.025	1.72 (4.44–1.72)	0.52 (1.18–0.52)	0.824
MTR (A2756G)									
AA	1.00 (ref.)	3.08 (2.07–4.57)		1.00 (ref.)	0.93 (0.68–1.28)		1.00 (ref.)	0.93 (1.40–0.93)	
AG	1.65 (1.05–2.59)	3.49 (2.26–5.40)		1.55 (1.03–2.33)	1.12 (0.77–1.61)		1.56 (2.40–1.56)	1.08 (1.70–1.08)	
GG	0.90 (0.27–2.94)	3.08 (1.45–6.53)	0.348	0.76 (0.27–2.14)	1.18 (0.54–2.56)	0.798	1.24 (3.45–1.24)	0.85 (1.92–0.85)	0.285
MTRR (A66G)									
AA	1.00 (ref.)	2.04 (1.31–3.16)		1.00 (ref.)	1.21 (0.83–1.76)		1.00 (ref.)	0.73 (1.15–0.73)	
AG	0.70 (0.45–1.09)	2.31 (1.49–3.58)		1.53 (1.02–2.27)	0.90 (0.61–1.32)		0.75 (1.14–0.75)	0.83 (1.31–0.83)	
GG	0.46 (0.20–1.09)	2.60 (1.49–4.56)	0.015	0.88 (0.45–1.73)	1.18 (0.67–2.06)	0.212	1.41 (2.82–1.41)	0.58 (1.09–0.58)	0.907
TS VNTR									
Non-2R/non-2R	1.00 (ref.)	2.62 (1.78–3.85)		1.00 (ref.)	0.93 (0.69–1.26)		1.00 (ref.)	0.88 (1.32–0.88)	
2R/non-2R	0.84 (0.51–1.37)	2.52 (1.64–3.88)		1.01 (0.65–1.55)	0.85 (0.58–1.24)		1.02 (1.60–1.02)	0.80 (1.28–0.80)	
2R/2R	0.60 (0.15–2.36)	1.70 (0.64–4.53)	0.668	0.82 (0.19–3.53)	0.44 (0.17–1.12)	0.550	0.66 (2.62–0.66)	0.48 (1.29–0.48)	0.673

[a]ORs were adjusted for age, sex, smoking habit, drinking habit, body mass index, total energy intake, carotene intake, vitamin C intake, vitamin E intake, folate intake, multivitamin use and referral pattern to our hospital.
[b]Interaction was modeled as a product of smoking habit (0, non-smoker and 1, smoker), drinking habit (0, non-drinker and 1, drinker), folate intake in score (0, tertile 1 and 1, textile 2 + 3) and genotype in score.

Discussion

The present study showed a significant impact of *MTHFR* C677T and *MTHFR* A1298C in combination for risk of the most smoking related subtypes of lung cancer, squamous and small cell carcinomas. Moreover, this effect was prominent among heavy smokers. The *MTHFR* 677TT genotype was inversely associated with squamous/small cell carcinoma risk among heavy drinkers. In combination analysis of smoking, drinking and folate consumption, several potential gene–environment interactions were suggested, between (i) the *MTRR* A66G polymorphism and smoking and (ii) the *MTHFR* A1298C polymorphism and alcohol consumption.

High dietary intake of folate has been found to decrease the risk of lung cancer in several epidemiological studies (5–7). Although our result for folate did not reach statistically significance, the observed trend was accordant with other studies. Two small-sized clinical trials found folate and vitamin B_{12} supplementation to reverse atypia among patients with bronchial squamous metaplasia, a precursor of squamous cell carcinoma of the lung (30,31). One might therefore hypothesize a protective effect of folate on lung cancer, but there are also epidemiological studies providing no support for this concept (32–35). Considering the fact that functional polymorphisms in folate-related genes may contribute to alteration of folate metabolism (15), it is biologically plausible to hypothesize that the polymorphisms or the gene–environment interactions rather than the folate intake alone have the impact on lung cancer risk.

Hitherto, only a few studies have investigated associations between one-carbon metabolism-related gene polymorphisms and lung cancer risk. The *MTHFR* 677TT genotype has been reported to decrease risk of lung cancer in female Caucasians (20), but the results were inconsistent in other case–control studies (17,19). The *MTHFR* 1298CC and *MTRR* 66AG or GG genotypes were associated with significantly increased risk (20,21), whereas *MTR* and *TS* enhancer region polymorphisms in the Caucasians studies demonstrated no link (21,22). Our results of overall analysis added evidence for a null association in this controversial issue. However, of note in this study was the fact that *MTHFR* 677T and/or *MTHFR* 1298C alleles were associated with reduced risk of squamous/small cell carcinomas, especially among heavy smokers and drinkers. It has been shown that subjects with the *MTHFR* 677TT and *MTHFR* 1298CC genotypes have a reduction in enzyme activity compared with the wild-type homozygous, 677CC and 1298AA genotypes (36–38). This would lead to high 5,10-methylene THF concentrations, which may provide more one-carbon groups for thymidylate synthesis, thereby enhancing DNA synthesis and repair ability. Thus, it is biologically reasonable that individuals harboring the *MTHFR* 677T and *MTHFR* 1298C alleles among heavy smokers and drinkers have lower risk of squamous/small cell carcinoma development, given that carcinogenesis is strongly related with the accumulation of DNA damage. To our knowledge, this is the first indication of protective effects of combinations of *MTHFR* polymorphisms for this histologic subtype. These data provide support for the hypothesis of links between one-carbon metabolism and tobacco and alcohol influence on squamous/small cell carcinoma carcinogenesis. Regarding other body sites, our previous study on esophagus cancers, which are almost all squamous cell carcinomas in Japan, demonstrated that the *MTHFR* 677TT had the protective effects among heavy drinkers, consistent with the present study (39).

One difficulty exists in distinguishing effects of smoking and drinking on lung cancer risk. In the present study, of 33 heavy drinkers in squamous/small cell carcinoma cases, 24 (72.7%) cases were heavy smokers, so we may not conclude an independent protective effect of *MTHFR* 677TT genotypes among heavy drinkers, although adjustment for smoking habits was performed. On the other hand, all cases with squamous/small cell carcinomas were smokers except one and 60% (85/142) in this subtype were heavy smokers (40 pack-years or more). Alcohol drinking as well as tobacco smoking is considered to induce DNA damage and resultant modification of nucleotides (40,41). In addition, high intake of alcohol can lead to folate depletion

Table V. Impact of combination of smoking and drinking habit by folate intake and the polymorphisms on lung cancer risk

	Folate intake							
	Tertile 1 (139.5–274.5 μg/day)				Tertile 2 + 3 (274.6–1481.0 μg/day)			
	Non-smoker/non-drinker	Smoker/non-drinker or non-smoker/drinker	Smoker/drinker[a]	*P* interaction[c]	Non-smoker/non-drinker	Smoker/non-drinker or non-smoker/drinker	Smoker/drinke[a]	*P* interaction[c]
	ORs[b] (95% CIs)	ORs[b] (95% CIs)	ORs[b] (95% CIs)		ORs[b] (95% CIs)	ORs[b] (95% CIs)	ORs[b] (95% CIs)	
MTHFR (C677T)								
CC	1.00 (ref.)	1.33 (0.44–3.99)	1.60 (0.58–4.41)		1.00 (ref.)	1.82 (0.90–3.67)	2.89 (1.43–5.84)	
CT + TT	0.61 (0.19–1.98)	1.66 (0.62–4.48)	1.55 (0.58–4.18)	0.763	1.54 (0.83–2.89)	1.48 (0.79–2.80)	2.93 (1.50–5.73)	0.389
MTHFR (A1298C)								
AA	1.00 (ref.)	3.18 (1.20–8.42)	2.78 (1.06–7.27)		1.00 (ref.)	0.97 (0.57–1.65)	1.70 (0.97–3.00)	
AC + CC	1.68 (0.52–5.48)	1.63 (0.55–4.81)	2.33 (0.84–6.47)	0.701	0.56 (0.29–1.08)	1.04 (0.56–1.91)	2.04 (1.10–3.76)	0.078
MTR (A2756G)								
AA	1.00 (ref.)	2.13 (0.90–5.04)	1.95 (0.83–4.62)		1.00 (ref.)	1.11 (0.64–1.92)	2.74 (1.53–4.89)	
AG + GG	1.63 (0.46–5.72)	3.00 (1.12–8.00)	3.28 (1.34–8.02)	0.826	1.38 (0.74–2.55)	2.03 (1.09–3.77)	2.16 (1.15–4.05)	0.069
MTRR (A66G)								
AA + AG	1.00 (ref.)	2.17 (1.00–4.73)	2.27 (1.05–4.91)		1.00 (ref.)	1.14 (0.71–1.82)	1.98 (1.18–3.32)	
GG	3.96 (0.49–32.23)	3.19 (1.02–10.03)	2.77 (0.88–8.78)	0.384	0.20 (0.04–0.91)	0.67 (0.24–1.88)	2.34 (1.12–4.90)	0.023
TS VNTR								
Non-2R/non-2R	1.00 (ref.)	1.75 (0.74–4.16)	2.14 (0.92–4.96)		1.00 (ref.)	0.95 (0.56–1.60)	2.04 (1.17–3.55)	
2R/non-2R + 2R/2R	0.87 (0.24–3.07)	2.38 (0.92–6.18)	1.48 (0.57–3.82)	0.367	0.58 (0.28–1.20)	1.30 (0.71–2.39)	1.65(0.90–3.05)	0.769

[a]Subjects who are both smoker and drinker.
[b]ORs were adjusted for age, sex, body mass index, total energy intake, carotene intake, vitamin C intake, vitamin E intake, multivitamin use and referral pattern to our hospital.
[c]Interaction was modeled as a product of smoking/drinking habit (0, non-smoker/non-drinker; 1, smoker/non-drinker or drinker/non-smoker and 2, smoker/drinker) and genotype in score.

(42). Therefore, it is within expectation that the *MTHFR* 677T allele, associated with high 5,10-methylene THF concentrations, may have the potential to protect against squamous/small cell carcinomas in tobacco consumers drinking large amounts of alcohol.

It was previously reported that lung cancer risk is higher with the *MTRR* 66AG/GG genotypes than the *MTRR* 66AA genotype among former smokers, but this did not extend to never and current smokers (21). Here, interaction between this gene and smoking habit was observed. Furthermore, the *MTRR* GG genotype exhibited a protective effect in low-risk subjects (non-smokers/non-drinkers with adequate folate intake). Several cytogenetic biomarker studies suggested that some polymorphisms involved in metabolic activation/deactivation or in DNA repair have been expected to be of special importance in modulating tobacco and alcohol carcinogen effects (43). A recent study reported a positive association with the modulating effect of the *MTRR* polymorphism on micronucleus frequency in peripheral blood lymphocytes, one of the cytogenetic markers (44), which is probably to increase by smoking (45) and drinking (46). The higher micronucleus frequency recorded in *MTRR* 66GG genotype with respect to AG or AA genotype is suggestive of a role of this polymorphism in modulation of chromosome stability, so that the findings may be consistent with our results. Further studies on the underlying mechanisms of the *MTRR* polymorphism thus appear warranted.

We found an interaction between the drinking habit and *MTHFR* A1298C polymorphisms for lung cancer risk, with decreased risk among non-drinkers. A Caucasian study showed that the *MTHFR* 1298CC genotype elevated risk among both drinkers and non-drinkers but only in women (20). The *MTHFR* A1298C is associated with decreased enzymatic activity (37,38) and would be expected to exert a similar effect to *MTHFR* C677T, with mutant alleles more protective among drinkers (27,39). There is no clear biological explanation for our results, and we cannot rule out the possibility that our observations for *MTHFR* A1298C were due to chance. Replication in a future study is needed.

Several potential limitations of the present study warrant consideration. First, internal validity of this hospital-based study is a potential threat to causal inference. We used non-cancer patients at our hospital as controls, given the likelihood that our cases arose within this population base, but individuals selected randomly from our control population were earlier shown to be similar to the general population in terms of the exposure of interest (47). Equivalence in the genotype distribution for the *MTHFR* C677T polymorphism between our controls and the general population has also been reported (48). To account for variation between cases and controls, we adjusted for referral pattern to our hospital. Second, as with other case–control studies, this study may suffer from recall bias. Although the questionnaires were completed before the diagnosis in our hospital, in some cases, patients referred from other institutions might have known the diagnosis. Third, we used a self-administered questionnaire to evaluate the nutrients intake, including folate. Data obtained from FFQ may not reflect intake as accurately as those from other methods, such as biological markers. We could not find any association with intake of vitamin C and E or folate for lung cancer risk, contrasting with our previous demonstration using the same population of protective effects of vegetables and fruits (4). The estimation of consumption by FFQ may be one possible explanation for this apparent anomaly. However, the reproducibility and validity of the FFQ were acceptable (28). We could not consider consumption of folate from supplements in total consumption, but the proportion of user with folate supplement is very low in Japan (0.1%) (49). Lastly, the limited number of cases, especially in subanalysis, is another factor and replication of our findings in a larger study is warranted.

In conclusion, we observed significant associations between *MTHFR* C677T and combined *MTHFR* C677T/A1298C polymorphisms and squamous/small cell carcinoma risk among heavy smokers and drinkers. Moreover, interactions between *MTRR* polymorphisms and smoking as well as the *MTHFR* A1298C polymorphism and alcohol consumption were also suggested. Our results thus support the hypothesis that folate metabolism-related gene polymorphisms may play a role in the genesis of lung cancer in combination with environmental factors. Replication in large epidemiological studies as well as studies of the mechanisms of the metabolisms is to be recommended.

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Genomic profiling of malignant pleural mesothelioma with array-based comparative genomic hybridization shows frequent non-random chromosomal alteration regions including *JUN* amplification on 1p32

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Genome-wide array-based comparative genomic hybridization analysis of malignant pleural mesotheliomas (MPM) was carried out to identify regions that display DNA copy number alterations. Seventeen primary tumors and nine cell lines derived from 22 individuals were studied, some of them originating from the same patients. Regions of genomic aberrations observed in >20% of individuals were 1q, 5p, 7p, 8q24 and 20p with gains, and 1p36.33, 1p36.1, 1p21.3, 3p21.3, 4q22, 4q34-qter, 6q25, 9p21.3, 10p, 13q33.2, 14q32.13, 18q and 22q with losses. Two regions at 1p32.1 and 11q22 showed a high copy gain. The 1p32.1 region contained a protooncogene, *JUN*, and we further demonstrated overexpression of *JUN* with real-time polymerase chain reaction analysis. As MPM cell lines did not overexpress *JUN*, our findings suggested that induction of *JUN* expression was involved in the development of MPM cells *in vivo*, which also might result in gene amplification in a subset of MPM. Meanwhile, the most frequent alteration was the 9p21.3 deletion, which includes the $p16^{INK4a}/p14^{ARF}$ locus. With polymerase chain reaction analysis, we determined the extent of the homozygous deletion regions of the $p16^{INK4a}/p14^{ARF}$ locus in MPM cell lines, which indicated that the deletion regions varied among cell lines. Our results with array comparative genomic hybridization analysis provide new insights into the genetic background of MPM, and also give some clues to develop a new molecular target therapy for MPM. (*Cancer Sci* 2007; 98: 438–446)

MPM, a highly lethal neoplasm of the serosal lining of the pleural cavity, is thought to develop from superficial mesothelial cells.[1] In up to 80% of patients, MPM occurs within about 30 years of exposure to asbestos.[2–4] The incidence of MPM is expected to increase dramatically over the next few decades. It has been estimated that 250 000 people will die of MPM in Europe in the next three decades, and 2500–3000 new cases are diagnosed each year in the USA.[5,6] In Japan, a recent report has shown that there will be approximately 100 000 deaths due to MPM in the next 40 years using an age-cohort model.[7] Survival of patients with MPM is very poor, with a median survival of 7–11 months after diagnosis, especially in advanced-stage patients, regardless of a recent advancement in chemotherapeutic modalities that combines cisplatin and antifolate.[8–10]

The long latency period between asbestos exposure and tumor development implies that multiple, and likely diverse, genetic changes are required for the malignant transformation of mesothelial cells. Many studies have been conducted to determine underlying key genetic and epigenetic events responsible for the development of MPM, some of which may be directly caused by asbestos fibers. Traditional karyotype analysis using primary samples or cell lines uncovered multiple non-random chromosomal abnormalities that are frequently detected in most human MPM specimens, which include chromosomes 1p, 3p, 6q, 9p and 22q.[11–18] Subsequent studies of such common regions with allele loss, which indicate the sites of TSG, have identified the target genes of MPM, including $p16^{INK4a}/p14^{ARF}$ on chromosome 9p21 and *NF2* at 22q. The $p16^{INK4a}/p14^{ARF}$ gene, one of the most frequently mutated TSG of human malignancies, has been shown to be inactivated in ~90% of MPM, with most cases being targeted by homozygous deletion.[19,20] The *NF2* gene at the 22q12 locus, which is responsible for a familiar cancer syndrome of neurofibromatosis type II, has also been shown to be inactivated in 40–50% of MPM, mainly with nonsense mutation or homozygous deletion.[21,22] In contrast, the *p53* gene, another of the most frequently mutated TSG in human malignancies, is only occasionally mutated in MPM, with approximately 25% of MPM specimens being inactivated.[23,24] Meanwhile, MPM does not show frequent mutation of known protooncogenes including *KRAS*, *NRAS* and *EGFR*.[25–28] Thus, it has been suggested that there are other yet unidentified TSG or protooncogenes responsible for the development of MPM. Recently, a CGH technique introduced to search for additional genes that are potentially involved in MPM biology has identified other regions with alterations, including 1q, 4q, 5p, 6p, 7p, 8p, 8q, 10p13-pter, 13q, 14q, 15q, 17p12-pter, 17q and 20.[29–34]

In the present study, we carried out array CGH analysis with 17 resected MPM samples (from 16 patients) and nine MPM cell lines from a total of 22 individuals. We confirmed the same chromosomal alterations as described before in the literature and further identified new regions such as 8q24 and 13q33.2. We also identified high copy gain at 1p32, which includes the *JUN* protooncogene. The present study provides new insights

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Abbreviations: BAC, bacterial artificial chromosome; CGH, comparative genomic hybridization; GAPDH, glyceraldehyde-3-phosphate dehydrogenase; MPM, malignant pleural mesothelioma; PAC, P-1 derived artificial chromosome; PCR, polymerase chain reaction; RT-PCR, reverse transcription–polymerase chain reaction; STR, short tandem repeat; TSG, tumor suppressor gene.

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Table 1. Summary of malignant pleural mesotheliomas analyzed with array comparative genomic hybridization (CGH)

KD number†	Sex	Subtype	Asbestos exposure	Cell line	*p16^{INK4a}/p14^{ARF}*‡	*NF2*§	*JUN*
332	Male	Epithelioid	+		HL	(+)	No Amp
355	Male	Epithelioid	–			(+)	No Amp
471	Male	Epithelioid	Unknown		HL	(+)	No Amp
476	Male	Biphasic	–	Y-MESO-8 A, -8D	HD	+	No Amp
905	Male	Epithelioid	Unknown		HL	del(533–537)	No Amp
977	Male	Epithelioid	Unknown			(+)	No Amp
1032	Male	Biphasic	+			(+)	No Amp
1033	Male	Epithelioid	+			(+)	Amp
1038	Male	Epithelioid	+			(+)	No Amp
1039	Male	Duciduoid	+			(+)	Amp
1041	Male	Duciduoid	+		L	(+)	Amp
1043	Female	Epithelioid	+			del(468–479)	No Amp
1044	Male	Epithelioid	–		L	(+)	No Amp
1045	Male	Epithelioid	–		L	(+)	No Amp
1046	Male	Biphasic	+		L	(+)	No Amp
1048	Male	Epithelioid	+	Y-MESO-9	HD	del(527–528)	No Amp
1049	Male	Epithelioid	+			(+)	No Amp
	Female	Epithelioid	–	Y-MESO-12	HD	+	No Amp
	Female	Epithelioid	–	ACC-MESO-1	HD	Q389X	No Amp
	Male	Epithelioid	+	ACC-MESO-4	HD	+	No Amp
	Male	Unknown	Unknown	NCI-H28	HD	[+]	No Amp
	Male	Unknown	Unknown	NCI-H2052	HD	R341X	No Amp
	Unknown	Unknown	Unknown	MSTO-211H	HD	[+]	No Amp

†KD Number indicates primary tumors available for array CGH analysis. Two primary tumors were obtained from the same patient at surgical resection (KD1039) and autopsy (KD1041). ‡*p16^{INK4a}/p14^{ARF}* status was indicated as follows: HL, high-level loss; L, loss; HD, homozygous deletion (detected in cell lines). § +, No point mutation was detected with PCR sequencing analysis of exons 1–17 covering the entire open reading frame of *NF2*, and homozygous deletion was not detected in the corresponding cell line; (+), no point mutation was detected in exons 1–17, but homozygous deletion was not determined due to possible contamination of non-cancerous DNA; [+], undetectable point mutation for exons 2, 5, 7, 8, 9, 10, 11 and 12. Data of *p16^{INK4a}/p14^{ARF}* and/or *NF2* of Y-MESO-8A, Y-MESO-8D, ACC-MESO-1, ACC-MESO-4, NCI-H28, H2052 and MSTO-211H referred to Sekido *et al.* and Usami *et al.*[21,28] Amp, amplification.

into the genetic background of MPM, and also gives some clues to developing a new molecular target therapy for MPM.

Materials and Methods

Cell lines and tumor specimens. Twelve MPM cell lines and one non-malignant mesothelial cell line (MeT-5A) were used. ACC-MESO-1, ACC-MESO-4, Y-MESO-8A, Y-MESO-8D, Y-MESO-9 and Y-MESO-12 were established in our laboratory,[28] whereas NCI-H28 (CRL-5820), NCI-H2052 (CRL-5915), NCI-H2373 (CRL-5943), MSTO-211H (CRL-2081) and MeT-5 A (CRL-9444) were purchased from the American Type Culture Collection (Rockville, MD, USA). NCI-H290 and NCI-H513 were gifts from Dr Adi F. Gazdar. All MPM cell lines were maintained in RPMI-1640 medium (Sigma-Aldrich, Irvine, UK) supplemented with 10% fetal bovine serum (Invitrogen, Carlsbad, CA, USA) and 1× antibiotic–antimycotic (Invitrogen) at 37°C in a humidified incubator with 5% CO_2. MeT-5 A was cultured according to the instructions of the American Type Culture Collection. Nineteen MPM samples from 18 Japanese patients were obtained at Aichi Cancer Center Hospital, Nagoya University Hospital, Japanese Red Cross Nagoya First Hospital, Nagoya Second Red Cross Hospital and Kasugai City Hospital (KD332, KD355, KD471, KD476, KD905, KD977, KD1032, KD1033, KD1038, KD1039, KD1041, KD1042, KD1043, KD1044, KD1045, KD1046, KD1048, KD1049 and KD1050; of these, KD1039 and KD1041 originated from the same patient at surgery and autopsy, respectively). MPM samples and clinical data were collected after obtaining appropriate institutional review board approval and written informed consent from all patients. To confirm that there was no cross-contamination of clinical samples and cell lines, the uniqueness or identity of MPM tissues and established cell lines were evaluated by analysis of STR polymorphisms using the AmpFLSTR Identifier Kit (Applied Biosystems, Foster City, CA, USA), including the 16 STR loci D8S1179, D21S11, D7S820, CSF1PO, D3S1358, TH01, D13S317, D16S539, D2S1338, D19S433, vWA, TPOX, D18S51, Amelogenin, D5S818, and FGA. Primary tumors and cell lines used in the present study are summarized in Table 1.

Preparation of DNA and RNA. Genomic DNA was extracted using a standard phenol–chloroform method.[35] Normal DNA was prepared from peripheral blood of healthy male donors and non-cancerous lung tissue of the patients. Total RNA was prepared using TRIzol reagent (Invitrogen) according to the manufacturer's protocol. DNase treatment was carried out on columns during RNA purification using an RNase-Free DNase Set (Qiagen, Germantown, MD, USA). Random-primed, first-strand cDNA was synthesized from 2 μg total RNA using Superscript II according to the manufacturer's instructions (Invitrogen).

Genome-wide array-based CGH. A genome-wide scanning array with 2304 BAC and PAC clones covering the whole human genome at a resolution of roughly 1.3 Mb was used as described previously.[36] In brief, clones were isolated from bacterial cultures containing the requisite antibiotics and extracted using a Plasmid Mini-kit (Qiagen). The location of all clones used for the array CGH was confirmed by standard fluorescence *in situ* hybridization analysis. BAC and PAC clones were amplified using degenerate oligonucleotide-primed PCR and spotted on glass slides. DNA preparation from cells, labeling, hybridization and scanning analysis were carried out as described previously[37] with minor modifications.[36,38,39] The data obtained were processed to detect chromosomal imbalances as described.[40]

Southern blot analysis. Genomic DNA from patient samples (7 μg) was digested with *Eco*RI restriction enzyme, electrophoresed,

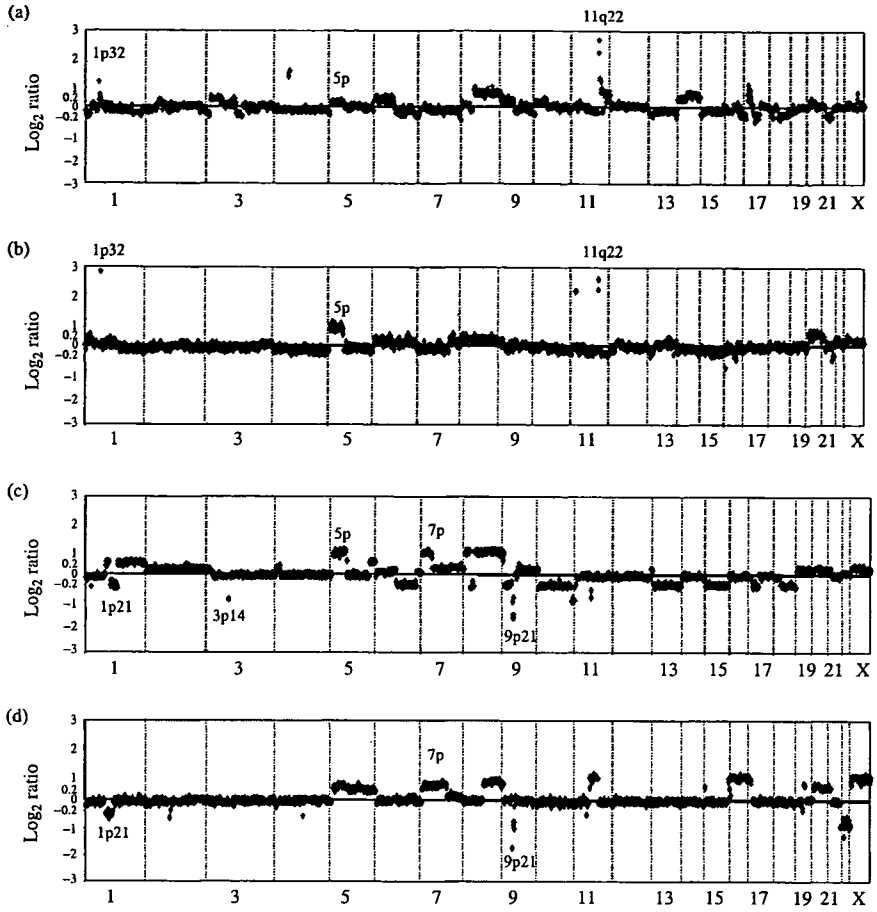

Fig. 1. Array comparative genomic hybridization profile of malignant pleural mesothelioma from three primary tumors and one cell line. Log_2 ratios are plotted for all clones based on chromosome position, with vertical dotted lines showing separation of the chromosome. Clones are ordered from chromosomes 1–22 and X within each chromosome on the basis of the Sanger Center Mapping Position, July 2004 version. (a) KD1033 sample shows chromosomal gain of 1p32.1-p32.3, 2p16, 3p22.2-pter, 3p12, 4q12, 5p, 6pter-q14.1, 8q, 9p, 10p, 11q22.1-q22.3, 11q23.3-qter, 14, 17p12-pter and 20p11.21-p12, and loss of 1p36.13-pter, 1q32-q42, 2q37.1-qter, 3q11-q13.31, 4q34.3-qter, 6q14.3-q21, 6q25-qter, 7q35-qter, 9q34.12-qter, 13q12.11-q13.3, 13q34, 16q23-qter, 17q11.2-qter, 18p, 18q12.2-qter and 21qcen-q22.2. (b) KD1041 primary sample shows chromosomal gain of 1p36.13-p36.32, 1p32.1, 5p, 6p22-pter, 6p12-p21.1, 8, 11p15.2-p15.3, 11q22.1, 20, 22q12-q13.2 and X, and loss of 3p21.31, 4q, 5q35.1-qter, 9p21.3, 11q23-qter, 13q12, 13q33.2, 15q22.3-qter, 16p13.2, 16q11-q12.2 and 21q22. (c) KD471 primary sample shows chromosomal gain of 1p22.2-p31.1, 1q, 2, 4p15-pter, 5p, 5q33.1-qter, 7, 8p21.1-pter, 8q, 9q, 12q24, 19 and 20, and loss of 1p36.31-p36.33, 1p36.13, 1p12-p22.1, 3p14.3-p21.31, 6q14-q25.1, 8p12-p21.1, 9p21.2-pter, 10, 11q12.1, 13, 15, 17p and 18q. (d) Y-MESO-12 cell line shows chromosomal gain of 5, 7pter-q21.3, 8q21-qter, 11qcen-q14.3, 15q11, 16, 19q13.2 and 20, and loss of 1p21-p31.1, 2p11, 4q22.1, 9p21.3, 11p12, 19p13.11 and 22.

and transferred to Hybond N+ (Amersham Biosciences, Piscataway, NJ, USA). Hybridization and washing were carried out using standard techniques.[35] The DNA probes were made by RT-PCR using normal lung cDNA. RT-PCR of *JUN* and *β-actin* were carried out using the primer sets: C-jun-S1, 5′-GACCTTATGGCT-ACAGTAACCC-3′ (sense) and C-jun-AS1, 5′-CTGCTCATCTG-TCACGTTCT-3′ (antisense); and B-Actin-S, 5′-CTGTGGCAT-CCACGAAACTA-3′ (sense) and B-Actin-AS, 5′-AGGAAAGACA-CCCACCTTGA-3′ (antisense).

Quantitative real-time PCR. The reaction mixture for real-time PCR using first-strand cDNA contained TaqMan universal PCR Master Mix (Applied Biosystems) and 200 nM of each primer, *JUN* (Hs 00277190_s1; Applied Biosystems) and *FOS* (Hs 00170630_m1). All real-time PCR assays were done in MicroAmp optical 96-well reaction plates on an ABI PRISM 7900 Sequence Detector System (Applied Biosystems) according to the manufacturer's instructions. For normalization between samples, PCR amplification of *GAPDH* (Hs 00266705_g1; Applied Biosystems) was included for each sample at each run. Fluorescence measurements and melting curve analyses were carried out using SDS 2.1 software (Applied Biosystems). The relative quantification of gene expression was computed using the comparative threshold cycle method with a mathematical formula described previously, and results are shown as a fold induction of mRNA.[41] We classified them into high-level expresser of *JUN* or *FOS* (defined as >0.15 of *JUN* or *FOS* mRNA expression relative to *GAPDH* mRNA expression), middle-level expresser (defined as >0.025 but <0.15), and low-level expresser (defined as <0.025).

Deletion mapping of 9p21. Information on 16 microsatellite markers and one sequence-tagged site marker at 9p21 was searched, and their sequences were obtained from the Human Genome Database (GDB) and the Ensembl Genome Browser. Three primer sets for exons 1, 2 and 3 of $p16^{INK4a}$ were as described previously,[28] and the primer set of exon 1β of $p14^{ARF}$ was p14ARF-F, 5′-CACCTCTGGTGCAAAGGGC-3′ (sense) and p14ARF-R, 5′-CCTAGCCTGGGCTAGAGACG-3′ (antisense).

Mutation analysis of *NF2*. Mutation analysis of *NF2* was carried out by direct sequencing after PCR amplification of genomic DNA. Seventeen primer sets covering the entire coding region of *NF2* were described previously.[28]

Results

Genomic profiles and data analysis of MPM. Array CGH analysis was carried out using genomic DNA samples extracted from 19 MPM primary tumors and nine MPM cell lines (ACC-MESO-1, ACC-MESO-4, Y-MESO-8A, Y-MESO-8D, Y-MESO-9, Y-MESO-12, NCI-H28, NCI-H290 and MSTO-211H). Among 19 primary tumors, we did not detect any significant genomic alterations in two tumors, which was probably due to much contamination of genomic DNA from non-malignant cells, and we excluded these tumors for further analysis. Of the 26 MPM analyzed successfully, there were paired samples from the same individuals: the Y-MESO-8 A and Y-MESO-8D cell lines were established from the KD476 primary tumor, Y-MESO-9 was established from KD1048, and the other two primary tumors (KD1039 and KD1041) were obtained from the same patient at surgical resection and autopsy, respectively. Thus a total of 22 individual MPM were studied (Table 1). All of the clones on chromosome X were analyzed separately because of sex mismatching. Copy number changes were detected at high-resolution for genomes as a whole for primary tumor samples as well as cell lines. We defined regions of gain or amplification as $\log_2$ ratio > +0.2, and regions suggestive of heterozygous loss or deletion as $\log_2$ ratio < −0.2. Figure 1 shows representative data of the entire genomic profiles of three MPM primary tumors and one cell

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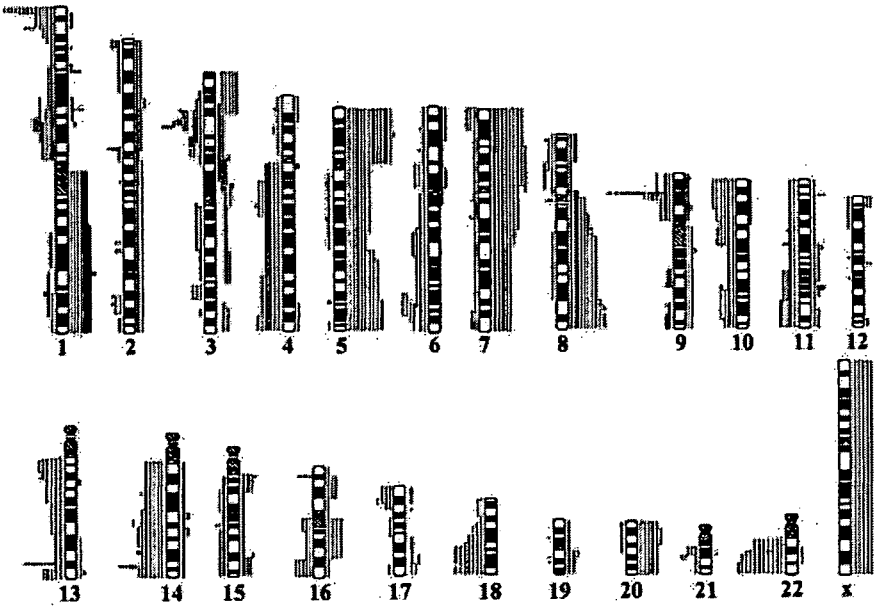

Fig. 2. Summary of chromosome imbalance detected in 17 malignant pleural mesothelioma patients (black lines) and nine cell lines (red lines). Regions of loss and gain are shown by vertical lines on the left (loss) and right (gain) sides of each ideogram. Regions of high-level amplification are presented by thick lines.

line from different individuals, with some shared altered regions being detected. For example, KD1033 (Fig. 1a) and KD1041 (Fig. 1b) showed shared regions including gain of 1p32.1, 5p, 8q, 11q22.1 and 20p and loss of 13q12 and 21q22. Figure 2 is a summary of chromosome imbalance detected in 17 MPM samples (black lines) and nine cell lines (red lines). Regions of high-level gain or amplification (defined as $\log_2$ ratio > +1.0) and those of homozygous loss or deletion (defined as $\log_2$ ratio < −1.0) are presented by thick lines. A summary of frequent chromosomal regions of gain and loss, and those of high-level copy gain or amplification, or homozygous loss or deletion detected in 17 MPM samples and nine cell lines is presented in Table 2. We also found that paired samples shared many chromosomal imbalances, although there were several different regions of gains and losses, or regions with relatively weak signals especially in the primary samples. The weak signals were thought to be due to contamination of non-malignant cell DNA (data not shown). Recurrent chromosomal imbalances found in at least six samples consisted of gain on chromosomes 1q (eight tumors/seven individuals), 5p (12/11), 7p (9/8), 8q24 (9/9), 20p (6/6) and loss on chromosomes 1p36.33 (13/13), 1p36.1 (7/7), 1p21.3 (7/6), 3p21.3 (10/8), 4q22 (7/6), 4q34-qter (6/6), 6q25 (7/6), 9p21.3 (16/16), 10p (6/5), 13q33.2 (11/9), 14q32.13 (13/11), 18q (7/6) and 22q (10/8).

High-level gain at 1p32.1 includes *JUN* protooncogene amplification. The array CGH analysis of 26 MPM revealed that 1p32.1 and 11q22.1 were two distinct regions with high-level gains, which were detected in at least two individual samples (Table 2). Interestingly, these high-level gains were observed simultaneously in the two individuals of KD1033 (Fig. 1a) and KD1041 (Fig. 1b). Another sample, KD1039, was also detected for 1p32.1 amplification (data not shown), and KD1039 and KD1041 were derived from the same patient, with the former at the initial surgical resection and the latter at autopsy. Whereas the KD1033 primary tumor showed a larger gain of five consecutive clones at 1p32.1 including the RP11-63G10 clone, KD1039 showed only a gain of the RP11-63G10 clone but not of the neighboring clones, and KD1041 showed only a gain of the two clones RP11-63G10 and RP11-363E22, with RP11-363E22 located toward the centromeric direction from RP11-63G10 1.9 MB apart (data not shown). Thus, the gain of RP11-63G10 seemed to be a very specific, common genetic event for these MPM, and this BAC clone was found to contain the protooncogene *JUN* (Table 2).

Because previous studies have suggested that asbestos fibers induce *JUN* expression in rat pleural mesothelial cells,[42] we studied the *JUN* status of MPM cells in further detail. We carried out Southern blot analysis with nine primary tumors and nine cell lines, and confirmed *JUN* high-level amplification in the three samples but not in the remaining 15 samples (Fig. 3a). To determine whether these MPM overexpress the transcripts of *JUN*, we carried out quantitative real-time PCR with 11 MPM samples available for RNA analysis together with seven MPM cell lines and one non-malignant mesothelial cell line, MeT-5 A. We found that KD1041, with high-level amplification of *JUN*, overexpressed mRNA of *JUN* (Fig. 3b). Interestingly, we noticed that there seemed to be three groups with distinct levels of *JUN* expression. We classified MPM into three groups according to the levels of *JUN* expression: high-level expresser (defined as >0.15) for three tumors (KD977, KD1041 and KD1044), middle-level expresser (defined as 0.025 < *JUN* < 0.15) for eight tumors (KD1032, KD1033, KD1045, KD1046, KD1048, KD1049, ACC-MESO-4 and H290), and low-level expresser (defined as <0.025) for seven tumors (KD471, KD476, ACC-MESO-1, Y-MESO-8A, Y-MESO-8D, H28 and MSTO-211H) and MeT-5 A. Among the seven MPM cell lines, ACC-MESO-4 and H290 were classified into middle-level expresser and the remaining five into low-level expressers. We also studied the *FOS* expression to determine whether *JUN* coexpresses with *FOS* in MPM cells (Fig. 3c). Most of the MPM cells classified into either high- or middle-level expresser of *JUN* simultaneously expressed *FOS* equal or greater than 0.025, and most expressers of both genes were primary tumors.

Alterations of *p16^{INK4a}/p14^{ARF}* at 9p21.3 and *NF2* at 22q12.2. We found frequent deletions of RP11-149I2 located at 9p21.3 in seven MPM samples and nine MPM cell lines, with five samples (two primary tumors and three cell lines) showing high-level loss. This BAC clone included *p16^{INK4a}/p14^{ARF}*, which is one of the most frequently mutated TSG in human malignancies, and we showed previously that *p16^{INK4a}/p14^{ARF}* was deleted in all MPM cell lines studied.[28] To determine whether the 9p21 deletion region in MPM extends further beyond the *p16^{INK4a}/p14^{ARF}* gene locus, which may indicate another target TSG of MPM in this region, we further carried out PCR analysis using multiple primer sets for comparison with locations of BAC and PAC clones on 9p21. Besides the nine MPM cell lines, another three MPM cell lines (NCI-H290, NCI-H513 and NCI-H2373) were also studied.

Table 2. **Chromosomal regions with frequent imbalances or high copy gain or loss detected in malignant pleural mesothelioma**

Alteration	Chromosomal region	No. patients (n = 17)	No. cell lines (n = 9)	No. individuals (n = 22)	Gene†	BAC/PAC‡
Gain						
	1p32.1§	3	0	2	*JUN*	RP11-63G10
	1q	4	4	7		
	5p	8	4	11	*CDH10*	RP11-116O11
	7p	5	4	8		
	8q24	4	5	9	*MYC*	RP1-80K22
	11q22.1§	2	0	2	*IAP*	RP11-864G5
	20p	3	3	6		
Loss						
	1p36.33	12	1	13	*KIT*	RP11-181G12
	1p36.1	4	3	7	*NM_018125*	RP11-473A10
	1p21.3	2	5	6	*RPL5*	RP4-716F6
	3p21.3	7	3	8	*PFKFB4*	RP5-1034C16
	4q22	2	5	6	*TMSL3*	RP11-309H6
	4q34-qter	3	3	6	*Q9P2F5*	RP11-739P1
	6q25	3	4	6	*PLEKHG1*	RP11-291C6
	9p21.3§	7	9	16	$p16^{INK4a}/p14^{ARF}$	RP11-149I2
	10p	2	4	5		
	13q33.2	7	4	9	*DAOA*	RP11-166E2
	14q32.13	8	5	11	*CHGA/ITPK1*	RP11-862G15
	18q	4	3	6	*MALT1*	RP11-4G8
	22q	7	3	8	*NF2*	RP1-76B20

†Representative genes are listed at each region when bacterial artificial chromosome (BAC) and P-1-derived artificial chromosome (PAC) clones of continuously ordered gain or loss of maximum overlapped clones were less than 10, when known protooncogenes or tumor suppressor genes shown to be involved in human malignancies were located, or when only a few genes were located in this region. ‡A representative BAC/PAC clone was listed when continuously ordered gain or loss of maximum overlapped region was less than 10 clones, and the clone at the mid-point of the overlapped region was chosen. §High copy gain or loss was observed.

After we confirmed homozygous deletions of exons 1, 2 and 3 of the $p16^{INK4a}$ gene and exon 1β of the $p14^{ARF}$ gene in all 12 (100%) MPM cell lines except MSTO-211H, which showed a partial retention of the gene, we used 16 microsatellite markers and one sequence site-tagged marker for the analysis (Fig. 4). For the telomeric direction, the INF-α cluster of genes was homozygously deleted in two cell lines but not in the remaining 10. For the centromeric direction, two cell lines (NCI-H290 and H2052) showed a larger deletion with consecutive losses at markers including D9S259, suggesting that these two cell lines had at least 4 Mb homozygous deletion. Meanwhile, four cell lines (Y-MES0-8A, -8D, NCI-H28 and H513) had a smaller homozygous deletion that was limited within D9S1749 and D9S790, suggesting that the maximum deletion size was less than 482 kb.

Finally, we studied any point mutations of the *NF2* gene in 17 primary tumors. After sequencing 17 exons covering the entire coding region of *NF2*, we found that three tumors had small deletions, all of which resulted in a frameshift mutation (Table 1). Because genomic DNA extracted from snap-frozen primary tumor tissues was used for the analysis, the existence of homozygous deletion was not determined due to possible contamination of non-cancerous DNA.

Discussion

In the present study, we analyzed 17 MPM primary tumors and nine MPM cell lines using array CGH and identified regions of genomic gain and loss. Regions of genomic aberrations observed in >20% of individuals were 1q, 5p, 7p, 8q24 and 20p with gains, and 1p36.33, 1p36.1, 1p21.3, 3p21.3, 4q22, 4q34-qter, 6q25, 9p21.3, 10p, 13q33.2, 14q32.13, 18q and 22q with losses. We confirmed the same chromosomal alterations as reported earlier by other groups and further identified high gain or amplification regions including 1p32, which harbors the *JUN* protooncogene. To our knowledge, our present study provides the first detailed array CGH data on chromosomal imbalances in MPM patient tumors and cell lines.

Traditional allelotyping and karyotype analyses revealed non-random chromosomal abnormalities including 1p, 3p, 4p15.1-p15.3, 4q25-q26, 4q33-q34, 6q, 9p, 14q11.1-q12, 14q23-q24 and 22q.[11–18,43,44] Subsequently, chromosomal CGH (also known as conventional CGH) has been carried out to detect more detailed abnormalities in MPM (Table 3). For example, Krismann *et al.* showed a total of 77 cases of MPM in the main histological subtypes (epithelioid type, sarcomatoid type and biphasic type) using chromosomal CGH.[34] They reviewed common gains at the chromosomal regions of 1q23/1q32, 7p14-p15, 8q22-q23 and 15q22-q25, and common losses at the chromosomal regions of 1p21, 3p21, 4p12-p13, 4q31-q32, 6q22, 9p21, 10p13-pter, 13q13-q14, 14q12-q24, 17p12-pter and 22q in all subtypes. In the present study with array CGH analysis, we also detected similar aberrations of multiple loci that have been found in previous studies.[29–35] These regions include gains of 1p32, 1q and 7p, and losses of 1p21, 9p21 and 22q. In addition to these regions, we have identified new regions such as 8q24 and 13q33.2, which had not been detected with chromosomal CGH analysis. The gain of 8q24 locus was detected by array CGH in nine cases (nine individuals) of these 26 samples. A single BAC, RP1-80K22, which includes the known protooncogene *MYC*, was located at the overlapped regions of 8q24 amplification. As a previous study showed a significant increase in signal strength of *MYC* in the mesothelioma tissues from an experimental animal model, compared with basal expression in non-neoplastic mesothelial cells, our findings also support the importance of *MYC* alteration in the development of MPM.[45]

Previous reports of chromosomal CGH analysis of MPM samples identified the region of gain at 1p32, although a specific candidate target gene was not referred to in detail.[34,46] Using array CGH, we found that a single BAC clone, RP11-63G10,

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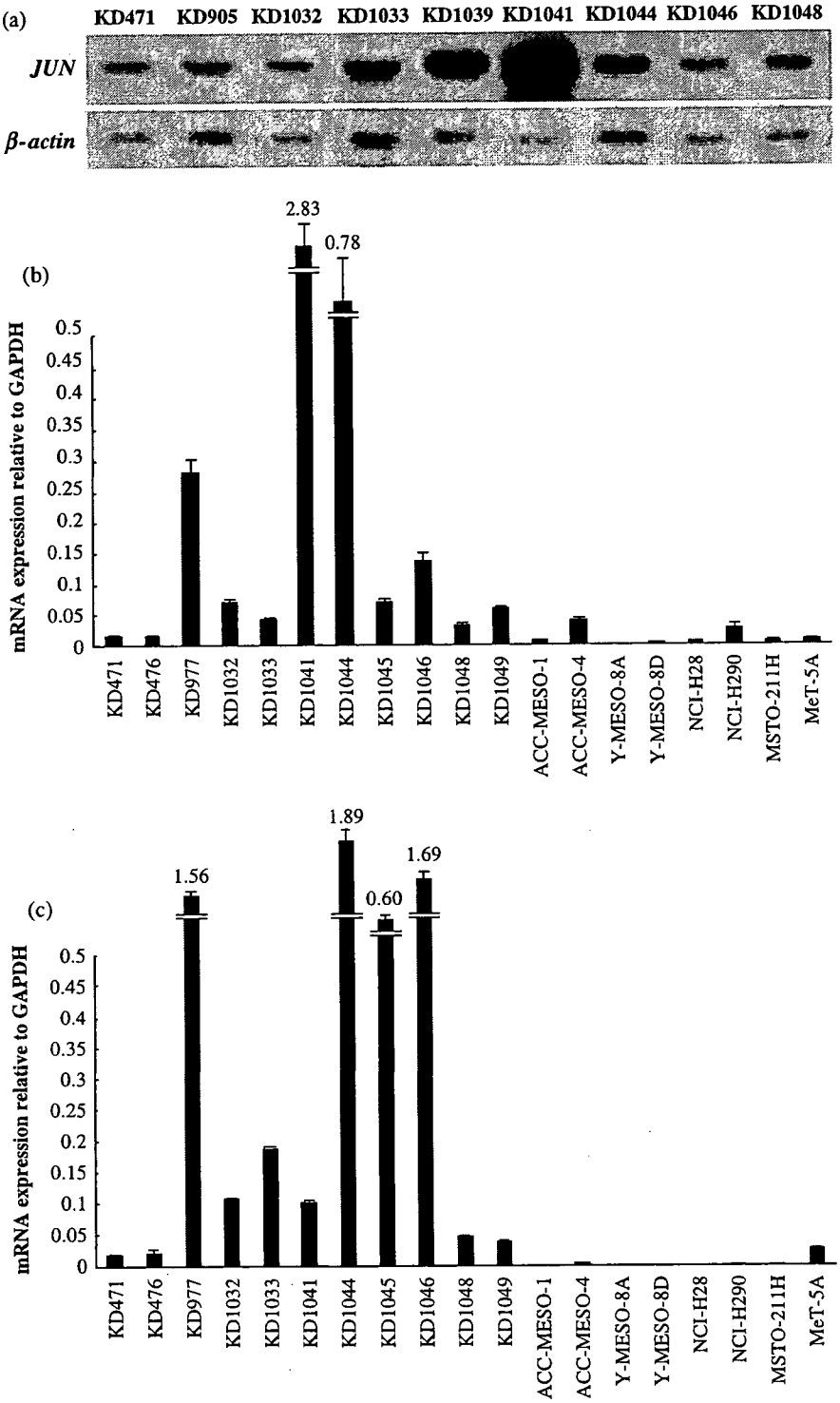

Fig. 3. *JUN* amplification at 1p32.1 and expression of *JUN* and *FOS* messages in malignant pleural mesothelioma. (a) Southern blot analysis of *JUN*. Each lane was loaded with 7 μg genomic DNA from MPM samples. Southern blot shows high-level amplification of *JUN* in KD1039 and KD1041 and low-level amplification in KD1033. (b,c) Diagrammatic presentation of quantitative real-time polymerase chain reaction data for (b) JUN and (c) FOS mRNA from 11 primary samples, seven MPM cell lines and MeT-5 A. The results were averages of at least three independent experiments with error bars showing standard deviations. MPM were classified into three groups of JUN status expression: high-level expresser (defined as >0.15) for three tumors (KD977, KD1041 and KD1044), middle-level expresser (defined as 0.025 < *JUN* < 0.15) for eight tumors (KD1032, KD1033, KD1045, KD1046, KD1048, KD1049, ACC-MESO-4 and H290), and low-level expresser (defined as <0.025) for the remaining seven tumors and MeT-5A. MPM were also classified into three groups according to *FOS* expression status: high-level expresser (defined as >0.15) for five tumors (KD977, KD1033, KD1044, KD1045 and KD1046), middle-level expresser (defined as 0.025 < *FOS* < 0.15) for four tumors (KD1032, KD1041, KD1048 and KD1049) and MeT-5 A, and low-level expresser (defined as <0.025) for the remaining nine tumors.

detected the region of gain at 1p32.1 in three tumors from two individuals. The RP11-63G10 clone was the only clone that showed overlapping at this region, and harbored only one known gene, the *JUN* protooncogene. Whereas KD1033 showed relatively wide-range amplification including five consecutive clones, KD1039 and KD1041 showed only RP11-63G10 amplification or with another neighbor clone for the latter (data not shown). It is noteworthy that KD1039 and KD1041 were from the same patient at surgical resection and autopsy, respectively, but the ranges of amplification of the *JUN* locus were slightly different. Furthermore, except for 1p32, these two samples also showed distinct regions of chromosomal alteration for each locus, including a gain at 13q34 for KD1039, and gains at 11p15.2 and 11q22.1 and a loss at 13q33.2 for KD1041 (data not shown). Although we confirmed the identity of these two samples with 16 STR repeats, it remains unclear whether the KD1041 cells originated from a subclonal cancer cell population that existed in the KD1039 tumor and acquired another chromosomal alteration during propagation.

JUN is a transcription factor and functions through homodimerization or heterodimerization with FOS to form the transcription factor AP-1, which can bind to the promoter region of

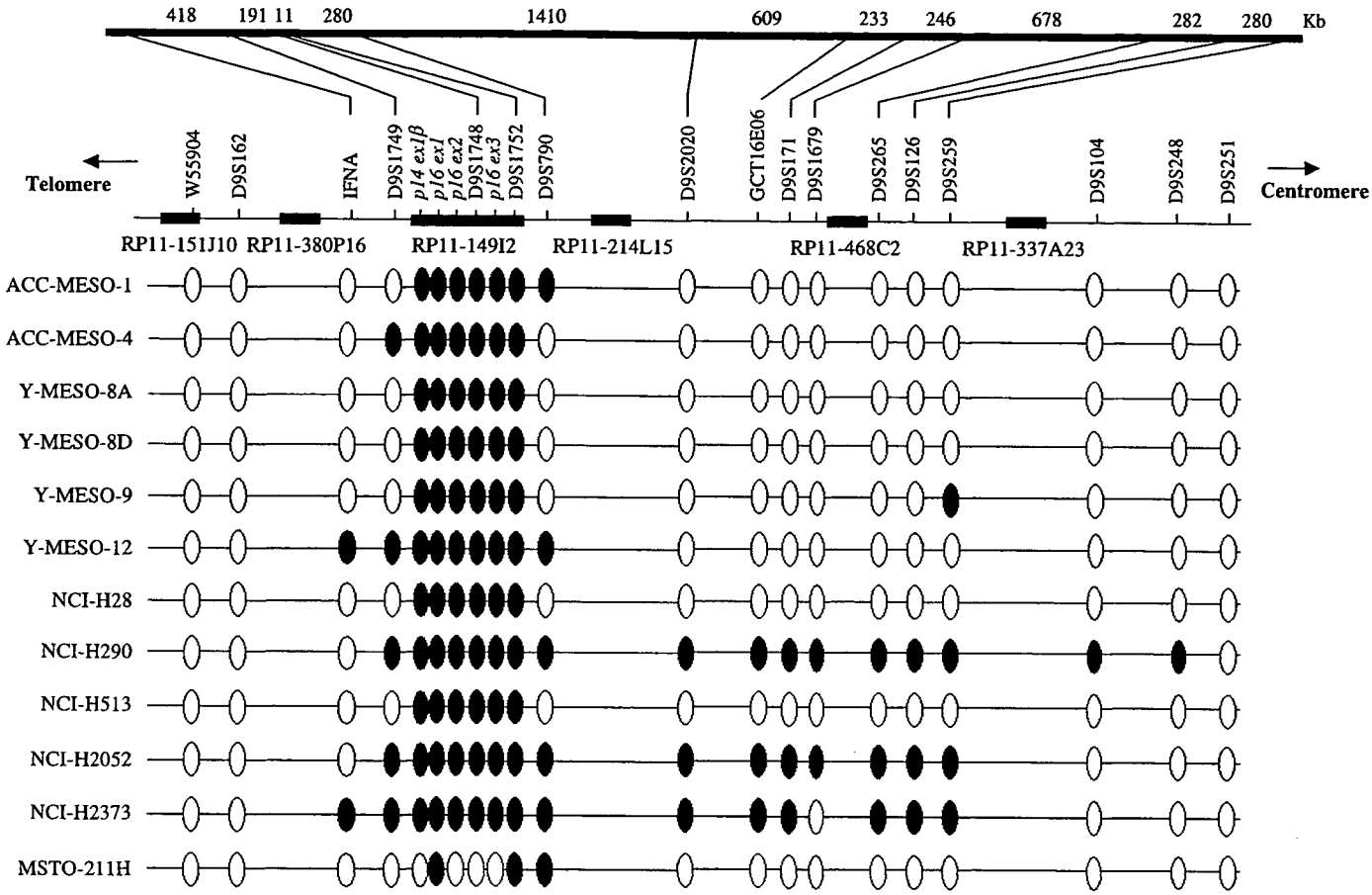

Fig. 4. Homozygous deletion map of the 9p21 region in 12 malignant pleural mesothelioma cell lines. Results of polymerase chain reaction analysis for each locus are shown by open ovals (retention) and closed ovals (homozygous deletion). Locations of genes and markers are according to those of the GDB Human Genome Database and Ensembl Genome Browser. Top bar shows the sizes between the selected markers proportionally: W55904 – (570 kb) – D9S162–(1.71 Mb) – IFNA – (418 kb) – D9S1749 – (191 kb) – D9S1748–(11 kb) – D9S1752 – (280 kb) – D9S790 – (1.41Mb) – D9S2020 – (609 kb) – GCT16E06 – (233 kb) – D9S171 – (246 kb) – D9S1679–(678 kb) – D9S265–(282 kb) – D9S126 – (280 kb) – D9S259 – (2.75 Mb) – D9S104 – (1.15 Mb) – D9S248 – (898 kb) – D9S251.

Table 3. Chromosomal regions with frequent imbalances shown in malignant pleural mesotheliomas from previous reports using chromosomal comparative genomic hybridization (CGH), and the current study using genome-wide array-based CGH

Authors	Year	Samples	Frequent gains	Frequent losses
Kivipensas *et al.*	1996	11	5p, 6p, 8q, 15q, 17q, 20	1p, 8p, 14q, 22q
Bjorkqvist *et al.*	1997	27	1cen-qter	4q31.1-qter, 6q22-q24, 9p21-pter, 13, 14q24-qter, 22q13
Bjorkqvist *et al.*	1998	34	7p, 15q	4q, 6q, 14q
Balsara *et al.*	1999	24	5p	1p12-p22, 6q25-qter, 9p21, 13q12-q14, 14q24-qter, 15q11.1-q15, 22q
Krismann *et al.*	2002	77	1q23/1q32, 7p14-p15, 8q22-q23, 15q22-q25	1p21, 3p21, 4p12-p13, 4q31-q32, 6q22, 9p21, 10p13-pter, 13q13-q14, 14q12-q24, 17p12-pter, 22q
Current study		26	1q, 5p, 7p, 8q24, 20p	1p36.33, 1p36.1, 1p21.3, 3p21.3, 4q22, 4q34-qter, 6q25, 9p21.3, 10p, 13q33.2, 14q32.13, 18q, 22q

intermediate genes involved in cell division and other cell functions.[47] Heintz *et al.* reported that both crocidolite and chrysotile asbestos caused increases in the expression of *JUN* and *FOS* in rat pleural mesothelial cells.[42] They demonstrated that, in contrast to phorbol 12-myristate 13-ester, which induced rapid and transient increases in *JUN* and *FOS* mRNA, asbestos caused 2–5-fold increases in *JUN* and *FOS* mRNA dose-dependently, which persisted for at least 24 h in mesothelial cells. They concluded that by activating the early response gene pathway, asbestos may induce chronic cell proliferation that subsequently contributes to carcinogenesis in lung and pleura. Thus, our findings of *JUN* amplification and overexpression detected in MPM tumors is very intriguing, and we also found that three tumors with *JUN* amplification were from patients with high-grade asbestos exposure. Interestingly, five of seven MPM cell lines were classified into low-level expressers of *JUN*, compared with three high-level and six middle-level expressers of the 11 primary tumors. This finding suggests that primary MPM tumor cells are

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continuously exposed to some stress to induce *JUN* transcription, and that *JUN* transcription is not necessarily induced in the established MPM cell line and MeT-5 A cells under usual tissue culture conditions, which may also indicate that the levels detected in MPM cell culture are of baseline *JUN* expression. Meanwhile, the analysis of *FOS* expression revealed that it was expressed simultaneously with *JUN* in most MPM cases, with high levels of expression of both genes detected mainly in the primary tumors, but not in cell cultures. These findings suggest the possibility that some surgical manipulations cause artificial induction of some genes, including early response genes,[48] which leads to the observation of predominant expression of these genes in the primary tumors. Nevertheless, because gene amplification of *JUN* was indeed identified in three MPM tumors, we think that there were some strong and persistent factors for *JUN* activation during the development of the MPM tumor cells.

JUN has been shown to be induced by other factors such as hypoxia. A recent immunohistochemical analysis detected expression of hypoxia-inducible factor 1α at focal regions in most MPM tumors but not in mesothelial cells, suggesting that hypoxic stress exists in primary MPM tumors.[49] Although the mechanisms and causes of amplification of genes such as *MYC* family members remain poorly understood, amplification of several other genes has been implicated as being induced by carcinogens and other stresses, such as amplification of the dihydrofolate reductase gene via methotrexate treatment.[50] Thus, we speculate that the chronic induction of *JUN* expression might have been induced by multiple stimuli, most importantly by asbestos fibers at the initial stage and possibly by hypoxia and other unidentified factors continuously, and that this might result in gene amplification of *JUN* in a subset of MPM cells during long latency.

Using array CGH, we found a region of loss at 9p21 in 16 tumors (16 individuals) that was covered by a single BAC clone, RP11-149I2, which included the $p16^{INK4a}/p14^{ARF}$ gene. It is well known that $p16^{INK4a}/p14^{ARF}$ is one of the most frequently deleted genes in many types of human cancers. Previous studies by other groups identified frequent alteration of $p16^{INK4a}/p14^{ARF}$ in most MPM, and we have also shown that $p16^{INK4a}/p14^{ARF}$ was deleted in all 10 MPM cell lines studied.[28] Although studies with simple PCR techniques reported homozygous deletion of $p16^{INK4a}/p14^{ARF}$ at a relatively lower frequency in MPM tissues than in cell lines, which may be due to contamination of a significant amount of normal stromal cells, we detected frequent deletion at 9p21.3 in seven MPM samples with array CGH. Furthermore, we determined the approximate lengths of deletion regions in 12 MPM cell lines, compared with the locations of DNA markers and BAC or PAC clones. We found that several cell lines showed a relatively small deletion with a maximum deletion size of 482 kb, whereas others showed at least a 4-Mb deletion size. Our findings of the $p16^{INK4a}/p14^{ARF}$ deletion in MPM seem consistent with other reports that the sizes of homozygous deletions vary individually in any given tissue type of malignancy.[51–53] Although it is very clear that $p16^{INK4a}/p14^{ARF}$ is the most important target TSG at the 9p21.3 region, other genes in this homozygous deletion region should also be studied to determine whether any of them play a role in the development of MPM.

Finally, the loss of 3p21.3 locus was detected by array CGH in 10 cases (eight individuals) of the 26 samples. One of the well-known TSG located at this region is *RASSF1A*, which is frequently inactivated by promoter hypermethylation in various types of human malignancies. The frequent hypermethylation of *RASSF1A* was also reported in MPM, which suggests that *RASSF1A* is a strong target TSG at 3p21 during the development of MPM.[54] Meanwhile, we also identified a homozygous deletion including *CTNNB1* (β-catenin) at 3p22.1 in the NCI-H28 cell line, and further demonstrated that the exogenously transfected *CTNNB1* gene inhibited the growth of NCI-H28 cells.[55,56] Thus, because several genes have been suggested as candidate TSG at the 3p21-22 region for various malignancies including MPM, further detailed analysis may be warranted to clarify the most important target TSG in this region for MPM.

To summarize, we subjected MPM samples to array CGH analysis and found genomic regions altered recurrently in MPM, including 1p32 *JUN* protooncogene amplification. Array CGH analysis can thus be expected to provide new insights into the genetic background of MPM and to offer some clues to developing a new molecular target therapy for this highly aggressive fatal tumor.

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PHASE II STUDIES

Phase II study of amrubicin in previously untreated patients with extensive-disease small cell lung cancer: West Japan Thoracic Oncology Group (WJTOG) study

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Summary *Purpose:* To evaluate the efficacy and safety of amrubicin, (+)-(7*S*, 9*S*)-9-acetyl-9-amino-7-[(2-deoxy-β-D-*erythro*-pentopyranosyl)oxy]-7,8,9,10-tetrahydro-6,11-dihydroxy-5,12-naphthacenedione hydrochloride, in previously untreated patients with extensive-disease small cell lung cancer (SCLC).

Patients and methods: A total of 35 previously untreated patients with extensive-disease SCLC were entered into the study. Amrubicin was given by daily intravenous infusion at 45 mg/m^2/day for 3 consecutive days, every 3 weeks. Unless there was tumor regression of 25% or greater after the first cycle, or 50% or greater after the second cycle, treatment was switched to salvage chemotherapy in combination with etoposide (100 mg/m^2, days 1, 2, and 3) and cisplatin (80 mg/m^2, day 1).

Results: Of the 35 patients entered, 33 were eligible and assessable for efficacy and toxicity. Of the 33 patients, 3 (9.1%) had a complete response (95% confidence interval [CI], 1.9–24.3%) and 22 had a partial response, for an overall response rate of 75.8% (95% CI, 57.7–88.9%). Median survival time was 11.7 months (95% CI, 9.9–15.3 months), and 1-year and 2-year survival rates were 48.5% and 20.2%, respectively. The most common toxicity was hematologic. Non-hematologic toxicity of grade 3 or 4 was only seen in 3 patients with anorexia (9.1%) and 1 patient with alopecia (3.0%). Salvage chemotherapy was administered to only 6 patients.

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Conclusion: Amrubicin was active for extensive-disease SCLC with acceptable toxicity. Further studies in combination with other agents for SCLC are warranted.

Keywords Amrubicin · Small cell lung cancer · Anthracycline · Previously untreated patients · Phase II study

Introduction

Small cell lung cancer (SCLC) is a major cause of cancer deaths and accounts for 15 to 20% of all lung cancers [1]. Although this cancer is initially highly responsive to chemotherapy, the vast majority of patients will ultimately relapse and die of recurrent disease within 2 years [2]. Recently, combination chemotherapy with irinotecan and cisplatin for extensive-disease SCLC produced more survival benefit than etoposide and cisplatin, the worldwide standard regimen since 1981 [3, 4]. Median survival time and 2-year survival rate of the standard regimen is 12.8 months and 19.5%, respectively. Clearly, new and more effective agents against SCLC are needed.

Amrubicin is a totally synthetic 9-aminoanthracycline, (+)-(7*S*, 9*S*)-9-acetyl-9-amino-7-[(2-deoxy-β-D-*erythro*-pentopyranosyl)oxy]-7, 8, 9, 10-tetrahydro-6, 11-dihydroxy-5,12-naphthacenedione hydrochloride, with a chemical structure similar to that of doxorubicin (Fig. 1) [5]. Amrubicin showed more potent antitumor activity than doxorubicin in several human tumor xenografts implanted in nude mice [6]. Acute toxicity of amrubicin is qualitatively similar to that of doxorubicin [7], however, amrubicin shows almost no delayed toxicity (e.g. cardiotoxicity) [8, 9].

Amrubicin is converted to an active metabolite, amrubicinol, by reduction of its C-13 ketone group to a hydroxy group. *In vitro* cytotoxic activity of amrubicinol was almost equipotent to that of doxorubicin and 20 to 220 times more potent than that of its parent compound, amrubicin [10]. Amrubicinol is considered to be closely associated with the efficacy and toxicity of amrubicin [11].

Despite their similarity in chemical structure, amrubicin has a different mode of action to doxorubicin [12]. Amrubicin and its active metabolite, amrubicinol, are inhibitors of DNA topoisomerase II. Amrubicin and amrubicinol exert cytotoxic effects by stabilizing topoisomerase II-mediated cleavable complexes, while doxorubicin does not inhibit this step of the catalytic cycle of topoisomerase II at concentrations for which it demonstrates cytotoxicity. Doxorubicin is a potent DNA intercalator, and its cytotoxicity is thought to be mainly due to this. Amrubicin and amrubicinol are about one-tenth weaker DNA intercalators than doxorubicin. Therefore, they are similar to etoposide in terms of inhibition of topoisomerase II by stabilizing the cleavable complexes, although etoposide does not show any DNA intercalating activity.

In a phase I–II study in patients with non-small cell lung cancer, amrubicin was administered as a 5-min intravenous infusion for 3 consecutive days [13]. The maximum tolerated dose (MTD) was 50 mg/m^2/day and the dose-limiting toxicities were leukopenia, neutropenia, thrombocytopenia, and gastrointestinal complications. The recommended dose for the phase II study was 45 mg/m^2/day for 3 consecutive days every 3 weeks.

Based on these experimental data and preliminary clinical reports indicating that amrubicin may be active against lung cancer, the West Japan Thoracic Oncology Group (WJTOG) evaluated it for use in SCLC. The WJTOG conducted a phase II study in previously untreated extensive-disease SCLC patients as a first-line therapy. Salvage chemotherapy with etoposide and cisplatin and an early cessation rule were set in place as precautionary measures.

Patients and methods

Eligibility criteria

Eligibility criteria included histologically or cytologically proven small cell lung cancer with extensive-disease defined as distant metastasis and/or disease involving the

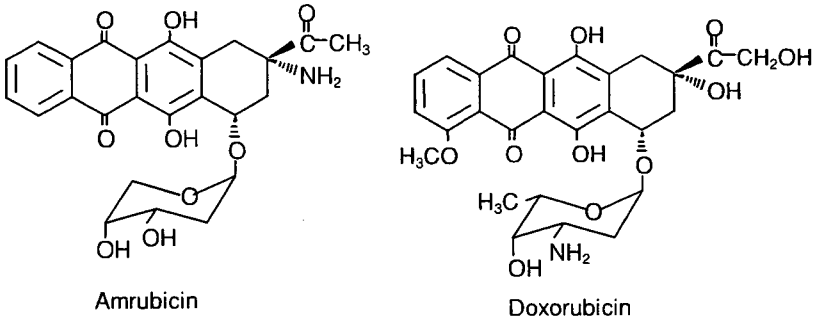

Fig. 1 Chemical structures of amrubicin and doxorubicin